SOME OBSERVATIONS ON THE
EGGS OF MOTHS AND CERTAIN ASPECTS
OF FIRST INSTAR LARVAL BEHAVIOR

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Note: Any figure numbers mentioned in this paper refer to egg photographs in the preceding paper (pp. 10(3): 215-247).

THE TIME REQUIRED FOR EGGS of the same moth species to hatch can vary tremendously with the temperature. It should always be stated under what conditions the eggs were kept and the month (or season) involved. (Outdoors: In the same habitat as the moth occurs? Indoors: Controlled conditions or fluctuating with the weather changes?) When kept unheated indoors, in the locality in which the species occurs, and subject to the normal day-night fluctuations of that locality, the great majority of species will hatch within a period of about 4 to 18 days, if they are not spp. that overwinter or aestivate as eggs. (In all instances where the number of days to hatch is mentioned for species, at the ends of the commentaries in the preceding paper, conditions under which those eggs were kept fit the last-described circumstances). Typical examples of variation in the number of days required for eggs of the same species to hatch (in the *same* locality but at different times of the year), are recorded in the commentaries for Fig. 6 and Fig. 31 of the preceding paper.

Some moths, especially univoltine species with autumn- or winter-emerging adults, in regions having a Mediterranean climate, will spend anywhere from 3 weeks to several months in the egg state. Of the 182 spp. of Australian "Macrolepidoptera" (representing 22 families) which I have reared from eggs in South Australia to date, I have encountered only one species which appears to approach overwintering in the egg stage here: *Epicoma tristis* Lewin (NOTODONTOIDEA-Thaumetopoeidae;

N.McF. code-number *Ta.8*). Even this one (possible) example needs more evidence to fully verify my suspicions. Some South Australian moths pass *part* of the winter as eggs (4 to 9 weeks), a not uncommon form of "partial overwintering" here. Most of these larvae hatch before the cold weather has ended, however.

In most species the adhesive which attaches the egg to the substrate is essentially colorless, although in some groups (Saturniidae, for example) it often colors the egg surface with irregular smudges of reddish-brown, or some other shade (depending on the species). The adhesive is generally more-or-less waterproof. This is imperative for the eggs of species having larvae that feed on the foliage of trees, shrubs, or high-climbing vines; such eggs could otherwise be easily washed from the foodplant leaves or stems during heavy rain. If this happened, the newly-hatched larvae would have little chance of finding their way back to the correct foodplant leaves during that short period after hatching when they are able to exist without food. Eggs lacking adhesive are often found among species having polyphagous larvae, root feeders, *or* feeders upon weeds, annuals, or other low plants that densely colonize areas of ground (bracken fern or *Pteridium*, for example). These moths often deposit or drop their eggs free (unattached); or the adhesive, if present, may be quite weak and the eggs readily become detached, which does not matter under these conditions. (The young larvae have little trouble finding their food near at hand when they hatch.) Such eggs are usually round, or at least approach a *spherical shape*. A South Australian example is the *Pteridium*-feeding ennomine geometrid, *Idiodes apicata* Gn. (Fig. 31).

Three fairly reliable indicators for the above (herbaceous, etc.) general foodplant categories are: Egg *"small"* for size of moth; *spherical* or nearly spherical egg shape; *lack of adhesive*, or only very weak adhesive. These clues can be of great assistance to anyone attempting to rear, from the egg, a species for which no foodplant was previously known. It eliminates most or all of the woody plants in the locality, and allows one to concentrate on the remaining flora. Exceptions, however, sometimes occur: In regions with the Mediterranean climatic pattern, where woody, sclerophyllous vegetation predominates (as in coastal southern and southwestern Australia), it is often typical of moths having larvae restricted to eating only (or primarily) the *young* and tender new leaves of woody sclerophyll trees or

shrubs, to produce eggs that are *also* small for the size of the moth. Such eggs may be of any shape, but are usually securely glued. Some examples of this latter condition, among southern Australian Geometridae, are: Oenochrominae—*Arhodia lasiocamparia* Gn. (Fig. 24); Ennominae—*Boarmia penthearia* Gn., *Capusa cuculloides* Felder (Fig. 36), *Cleora displicata* Walker (Fig. 34), *Gastrinopa xylistis* Lower, and *Melanodes anthracitaria* Gn. (Fig. 39). Eggs appearing "large", for the size of the moth—again, egg shape does not matter—more often than not imply feeders upon the foliage of *woody* plants, or the *mature* foliage of woody *sclerophyll* plants, or (occasionally) the leaves of grasses, especially the tougher perennial species. Newly-hatched larvae that cling with considerable or great tenacity are almost always feeders on (foliage of) woody plants, and they often have slow (or relatively slow) locomotion; this does not, of course, apply to *all* feeders on woody plants, but it has proven to be a very useful clue in numerous cases. I have repeatedly observed that species depositing eggs which look "small", for the size of the moth, frequently have larvae which grow exceptionally fast; even though they start out at a smaller size, they often surpass the growth-rate of other larvae which originally hatched from relatively larger eggs, and were larger at the start. These faster-growing larvae are usually feeders on "soft" vegetation (tender low-growing annuals, weeds, or the *new* leaves of woody plants.) "Excellent contrasting examples of this phenomenon are seen in Figs. 37 and 39 of the preceding paper. *L. habrocosma* is a *much* smaller moth than *M. anthracitaria*, yet compare the egg measurements of these two species! The former feeds *only* on the tough, old (sclerophyll) leaves of its foodplant; the latter *only* on the tender new leaves of its (sclerophyll) foodplant."

Moths possessing noticeably long and easily-extended ovipositors have the habit, more often than not, of ovipositing *into crevices*, under flakes of bark, into buds, or down between stems and sheaths of monocots, *etc.* Such eggs usually have thin or very easily-broken shells, and are often deposited in groups or masses, with no covering of scales or froth, etc. *Cleora displicata* Walk. (Fig. 34) is a good South Australian example. For success in obtaining eggs from such species in captivity, it is often worthwhile to provide rough paper towel, tightly folded fan-like (to create many crevices). Sometimes, a piece of paper tightly rumpled will do the same job. If they can be induced to

oviposit on paper, the eggs are more easily retrieved later, for study or photography. In some cases, however, rough or shredded bark or something closer to the texture of the actual substrate, as sought out by the moth in its habitat, must be provided in order to create the needed oviposition stimulus.

Interesting questions arise with reference to certain very "thin" or soft-shelled eggs, when the species involved deposit naked eggs in open or *exposed* situations—for example, on *Eucalyptus* leaf surfaces in the case of the Australian limacodid, *Pseudanapaea trigona* (Turner) (my Lm. 3). The entirely naked eggs of this moth (see also Fig. 4 and its commentary), are readily subject to desiccation if kept in certain types of containers, or if the leaves to which they were attached have been allowed to completely dry out. Perhaps, when on a *living* leaf surface, they may receive the required moisture-balance from the leaf stomata (during transpiration) immediately around and/or underneath the eggs(?) It is possible that among other Zygaenoidea (Limacodidae, Megalopygidae, or Zygaenidae, etc.), or among the Pyraloidea, there may be similar cases. Deeper investigation into this subject might produce some interesting results.

A phenomenon worth reporting, in connection with relative egg-sizes in different populations of a single species, has been noticed for one of the South Australian moths having a wide distribution that extends from coastal areas far into the arid interior. The moth involved is a notodontid, *Danima banksiae* (Lew.). (See also the commentary for Fig. 10.) Field-collected eggs of this species from a coastal population, 10 miles W. of Vivonne Bay, Kangaroo Island, South Australia—mid OCT. 1966—N. McFarland (my Nd.15B), had measurements as follows: 2.00-1.85 mm. (diameter) x 1.40-1.35mm. (height), based on a long series. Field-collected eggs of this species from a desert population, 72 miles S. of Kulgera (Northern Territory), *in* South Australia 25 May 67—N.McF. & T. Newbery (my Nd. 15D), had measurements as follows: 2.30-2.25 mm x 1.65 mm., based on 3 eggs only. The eggs of this latter (desert) population were notably so much larger than those of coastal populations (with which I was already familiar), that the size difference was immediately apparent, even in the field. Two perfect adults were reared from the 3 field-collected desert eggs, producing typical *D. banksiae,* hardly distinguishable (in size, maculation, or coloring) from coastal adults. The third was preserved as a last instar larva (in S.A. Museum, Adelaide—Nd.15D).

Other impressions gained (not enough data yet to clarify), concerning the relative egg sizes of coastal versus inland geometrids of certain related Australian genera and species, tend to further reinforce an idea that eggs of desert (or relatively dry area) populations are sometimes a little larger than eggs of the same species (or close relatives) having populations in coastal, more humid, or wetter localities. (See also remarks under Fig. 15, egg dimensions of *Cosmodes elegans.*) Further observations along these lines will probably uncover parallels in other regions.

Rain-hatched eggs: A number of South Australian geometrids (about 10 of the Ennominae and 2 of the Larentiinae which I have reared thus far) have what I shall refer to here as "rain-hatched eggs". The adults of these spp., without exception, emerge *between* mid March and late May (autumn to early winter), which marks the beginning of the heavy rains (and cooler weather) in coastal South Australia. The eggs of these species do not at first show any peculiarities, but, as the larvae develop inside and approach apparent readiness to hatch (the fully developed larvae being clearly visible through the more-or-less transparent shells) they persistently "refuse" to emerge until such time as water is applied to them. They will, in the more strict examples of this phenomenon (for example, the ennomine geometrid, *Mnesampela fucata*), shrivel up and die inside the eggs if ample moisture is not present when required to trigger emergence. Yet, if these eggs, when definitely *ready* to hatch, are watered, and the excess water is poured off, hatching will sometimes begin within 5 minutes of the wetting! (*Amelora amblopa* Guest—Ennominae; my G.143). If not that rapidly, they will almost invariably be out within 30 minutes to 3 hours, or in half a day at the most. Other examples of South Australian geometrids having rain-hatched eggs are: ENNOMINAE—*Amelora milvaria* Gn. (my G.114), *Chlenias* (*pachymela* Lower?) (G.106), *C. umbraticaria* Gn. (G.112), *Mnesampela lenaea* Meyr. (G.118), *Stathmorrhopa macroptila* Turner (G.99), *Stibaroma trigramma* Lower (G.104), *Thalaina angulosa* Walk. (G.100); LARENTIINAE — *Xanthorhoe heliacaria* (Gn.) (G.142).

There are a number of other South Australian geometrid species which appear to be on the borderline of this requirement. Examples are the *oenochromine, Cycloprorodes melanoxysta* (Meyr.) (my G.221), the desert *ennomine, Thalainodes macfarlandi* Wilson (G.180), the coastal *ennomine, Lophothalaina habrocosma* (Lower) (G.154), and the *larentiine, Xanthorhoe*

vicissata (Gn.) (G.92); also an *anthelid,* one of the *Pterolocera*
spp. (An.2A). Larvae of these species will mostly emerge from
their eggs sooner or later without the addition of moisture, but
wetting (or just increased humidity) definitely speeds up the
process, bringing out the whole batch more easily and uniformly,
within one or two days.

It is possible that dew may suffice to cause hatching in some
of these cases, under natural conditions outdoors, particularly
if the species involved is a feeder upon weeds, or other low
plants on the ground (?) (*X. heliacaria* might be an example
here). But, in dry jars or other containers indoors, neither dew
nor rain enters the picture.

Larvae in rain-hatched eggs are capable of waiting inside the
egg, fully ready to hatch, for many days (even weeks in some
cases) beyond the day when they *first* became ready to hatch,
and will do just that if kept dry. Exactly how long the larvae
of these species can remain alive like this I have not yet deter-
mined; it undoubtedly varies with the species. Awareness of
this peculiarity is a necessity for the successful hatching of any
such eggs in captivity. It is most important, however, that
moisture NOT to be applied until the young larvae can be seen,
fully-developed inside the egg shells, or the eggs (if enclosed in
a glass jar) are very likely to develop mould and/or fail to
hatch. *More than one* quick soaking (always followed by a slow
drying) is sometimes required to induce hatching, once the
correct time has arrived.

Although I have reared (from the egg) 28 Australian and 7
North American spp. of Geometrinae (Hemitheinae) to date,
I have not encountered a single example of rain-hatched eggs
in this subfamily.

Some advantages of the rain-hatched egg could be readily
imagined for species inhabiting regions where a long dry season
is finally broken by the first rains (the *dates* of which are rarely
predictable or consistent) of autumn or early winter. If the
larvae, ready to hatch, remain inside their eggs (where desicca-
tion is reduced to a minimum) until the arrival of a good rain,
the entire habitat then becomes moist (including the air), and
their chances for escaping desiccation while still small are
greatly increased. This would be all the more important when
a species also has strongly-developed first instar dispersal be-
havior, as it would increase the potential length of the survival-
period while the larvae are still wandering. Incidentally, all

species having rain-hatched eggs do *not* show first instar dispersal (nor vice versa), but some of them do.

First instar dispersal: A notable aspect of first instar larval behavior, which I shall refer to here as "first instar dispersal". is typical of a number of South Australian species, particularly in the GEOMETRIDAE, Subfamily *Ennominae*. I have also observed it in the NOTODONTIDAE (*Sorama biocolor* Walk.), developed to a modest degree, and highly developed in two genera (three species) of the THAUMETOPOEIDAE (examples = *Discophlebia catocalina* and *Oenosandra boisduvalii*). In the case of the GEOMETRIDAE, 14 spp. (13 ennomines and 1 oenochromine), out of the 112 spp. of Australian geometrids I have reared from eggs to date, have shown this behavior developed to a high degree. (It might be expected to crop up in a number of other Australian moth families as well, such as Psychidae and Lymantriidae, etc.). Twelve examples of South Australian geometrids demonstrating first instar larval dispersal are: OENOCHROMINAE—*Arhodia lasiocamparia* Gn. (my G.124); ENNOMINAE—*Boarmia penthearia* Gn. (G.211), *B. suasaria* Gn. (G.78), *Capusa cuculloides* Felder (G.105), *Cleora displicata* Walk. (G.165), *Ectropis excursaria* (Gn.) (G.75), *Stathmorrhopa eribola* (Guest) (G.90), *Gastrinopa xylistis* Lower (G.137), *Melanodes arthracitaria* Gn. (G.161), *Mnesampela fucata* (Felder) (G.109), *Stibaroma melanotoxa* Guest (G.81), and *S. trigramma* Lower (G.104).

Species characterized by first instar dispersal usually have most (or all) of the following features in common:

(1) The eggs often (but not always) appear "small" for the size of the moth.

(2) Large numbers of eggs may be deposited (when compared with other members of the same subfamily not having this behavior, or with other adults of similar size); yet these same moths are very often scarce species (or at least not particularly common) in the adult stage. In such species, the (probable) high mortality-rate, during the dispersal period, may, at least in part, explain some of the cases of apparent low numbers of *adults* among certain spp. feeding on abundant (or widespread) foodplants. Two South Australian examples representing this situation fairly well are *Capusa cuculloides* (Fig. 36) and *Melanodes anthracitaria* (Fig. 39.)

(3) The eggs are usually deposited very securely-glued, and in large masses or other group formations. The fact that the eggs of species having first instar dispersal are usually deposited in large masses or groups implies that some form of early dispersal would almost have to take place, *except* in those cases where the larvae are normally gregarious or at least semi-gregarious (for example the ennomine geometrid, *Mnesampela privata* (Gn.), and some of the thaumetopoeids, such as *Ochrogaster* spp., or the early instars of *Epicoma* and *Trichetra* spp.). There are some good explanations

for first instar dispersal among species having solitary larvae but depositing their eggs in large masses: These spp. often (not always) deposit their egg masses on the trunks or branches of trees, yet their larvae feed upon the leaves (often only the newer growth at the ends of branches), Therefore, the young larvae must crawl (disperse) actively upward and outward from the hatching-site, if they are ever to reach their food supply. Even when the masses are deposited on the foodplant leaves, these larvae would still be highly vulnerable to predation if they did not disperse soon after hatching. (Such species rarely, if ever, represent forms protected by aposematic colors). Under natural conditions the mortality-rate is almost certain to be extremely high during this active dispersal period. Winds could carry many of the young larvae (when dropping on silk) to other trees or shrubs, *not* necessarily always of acceptable foodplant species, and great numbers are probably taken by predators (ants, young spiders, etc.) during their early wanderings.

(4) These larvae, upon hatching, have a period (usually 2 to 5 days) of almost ceaseless lively wandering, during which time they show marked phototropism, and (depending on the species) are frequently inclined to drop or hang on silken threads. This habit of repeatedly dropping on silk tends to greatly increase the rate of dispersal under natural conditions, as the larvae of most of these species are in trees, thus being readily blown about by any wind or breezes.

(5) The foodplants of these species are nearly always *trees* (less often large shrubs). If the plant is sclerophyllous, usually only the tender new leaves are eaten; in such cases the larvae often appear to be *restricted* to the new growth and will refuse to eat older (tough) leaves, or will not grow well on them. The latter is by no means invariably true, but it applies more often than not. Three South Australian ennomine examples are *Capusa cuculloides, Gastinopa xylistis,* and *Melanodes anthracitaria;* these spp. feed only on the tender new growth of their foodplants if allowed their preference.

I have yet to see a single example of first instar dispersal behavior among the 28 Australian (or 7 North American) spp. of Geometrinae (Hemitheinae) reared from eggs to date. They are, in most cases, *highly* sedentary larvae right from the moment of hatching. Eggs of Geometrinae are usually deposited singly, or (at most) in twos and threes on the foodplant leaves; less often, in *short* curving rows, *small* "stacks", or irregular *small* clusters. Thus, in the Geometrinae, dispersal is usually accomplished entirely *by the female moth during oviposition.* The larvae hatch where they will be feeding, already dispersed, so there is no cause for additional dispersal to take place during first instar.

Species having first instar dispersal behavior can be difficult to start in captivity when rearing is attempted, but generally give no further trouble once they have *settled down* to begin feeding. During the dispersal period these newly-hatched larvae normally refuse to settle down on *any* plant to feed, *whether*

or not the correct foodplant is present. This can cause failure
to locate a suitable foodplant, (and thus failure in an attempted
rearing), if one does not recognize first instar dispersal behavior.
Careful attention to the following suggestions will lead to suc-
cess with the rearing of these species: (1) Do not draw any
firm conclusions as to *which* of the offered plants are supposedly
"unsuitable" until *after* the dispersal period is over. (2) Do not
fail to provide sheets of tissue or paper towel in the bottoms of
all foodplant sample-jars (into which the newly-hatched larvae
are placed). This allows the larvae to more readily untangle
themselves from their (often) abundant silken threads, on which
they may drop frequently, and to more quickly regain footing
among the plants whenever they drop on silk, or when they
crawl off onto the sides and bottom of the jar (which they will
do endlessly). It is imperative that many of the offered leaves
reach down to the *bottom* of the jar (contacting the towel), and
that other leaves extend higher up in the jar (with ample space
between them), in order to allow those larvae that have begun
to settle to partially isolate themselves from others still actively
crawling; otherwise, the latter will constantly disturb those
trying to start feeding, which will in turn set them to wandering
all over again! (3) Place their foodplant sample-jars inside a
totally dark cupboard, at a cool or mild temperature (not over
70 F., if possible). When in a partially lighted situation, their
positive phototropism causes them to constantly migrate toward
that side of the jar facing the strongest light source, which is
also the side on which maximum condensation will occur. They
often became trapped in this moisture, and (at the same time)
will fail to uniformly scatter through the offered plant samples,
thus "missing" many of the samples until it is too late. (Any
becoming stuck in condensation are easily lifted off by means
of a small watercolor brush, and can be replaced among the
plants). The darkened-cupboard-technique, however, overcomes
these difficulties with great efficiency. (4) Alternatively, they
can be left in a lighted situation *provided* that the jar is regularly
(at least 2 or 3 times daily) turned halfway around, thereby
forcing the larvae to crawl straight back through the plant
samples as they head (again) toward the brightest light. (Even-
tually they will tire of this, and begin to slow down). Under all
circumstances, the piece of paper towel in the bottom is of vital
importance. Also, they must be frequently and critically checked
during this wandering period, so that *any* impending trouble
will be detected early, and necessary preventive action can be

taken. With many newly-hatched larvae, only a few *hours* under wrong conditions can spell the finish of a proposed rearing project.

Due to the condensation problem, it might be imagined that a screen cage (or semi-cage) would be more suitable for starting small larvae, but this is definitely not so in the case of species having first instar dispersal, *unless* the cage is exceptionally well made (absolutely no tiny cracks), is of a relatively small size, and is made from a very fine mesh material (for example, "Nylon Tule", available at 60c per yard in certain Australian department stores). This will prevent the escape of active small larvae, while providing limited ventilation. (Only *limited* ventilation is desirable). Desiccation of larvae and food-plants is a common problem in a well-ventilated cage. They must be kept reasonably cool, and may need to be frequently sprayed with a fine water-mist under some conditions. Such problems do not arise when using the *closed-jar* technique for starting small larvae (McFarland, 1964, 1965; ref. in preceding paper). Larvae of even the most disease-prone species seem to be amazingly disease-resistant when small (first and second instars±), and are able to tolerate much higher humidity then. By about third instar they are usually ready to transfer from jars to partially ventilated cages, *without* any risk of loss through escape or desiccation.